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Ectoparasite Presence in Select Northcentral Kansas Bat Species

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ECTOPARASITE PRESENCE

IN SELECT NORTHCENTRAL KANSAS BAT SPECIES

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PREFACE

This thesis is written in the style of the Journal of Mammalogy, to which a portion will be submitted for publication.

ABSTRACT

Working with other graduate students on a grant given to Fort Hays State University, from the Kansas Department of Wildlife, Parks and Tourism, I looked at presence and species of ectoparasites on bat species. The main goal of our grant was to quantify and qualify the status of the northern myotis (*Myotis septentrionalis*) in the state of Kansas, and to record data on any bycatch. I worked on our grant in the summer field seasons of 2016 and 2017, May to October, as described by the Indiana bat protocol.

Bats were captured by using mist nets set over ponds, small streams, and rivers in northcentral Kansas. I chose sites by using a combination of historical and acoustic data. I mist netted 61 nights in the field season of 2016, and 47 nights in the field season of 2017.

Over the field seasons of 2016 and 2017, I captured the following bat species: *Eptesicus fuscus, Lasiurus borealis*, *Lasiurus cinereus*, *Myotis septentrionalis*, *Nycticeius humeralis*, and *Perimyotis subflavus*. Only the evening bat, *N. humeralis*, was captured in numbers large enough to run statistical analyses. I compared the presence of ectoparasites between adults and juveniles, males and females, male reproductive status, and female reproductive status. When compared, adults had a significantly lower presence of ectoparasites than juveniles did ($X^2 = 47.38$, d.f. = 3, $p = 0.00001$). Only 33% of adult *N. humeralis* had ectoparasites, while 76% of juveniles had ectoparasites. Males had 72% ectoparasite presence while females only had 41% ectoparasite presence ($X^2 = 15.03$, d.f. = 3, $p = 0.01792$). When males were compared based on their

reproductive status there was no statistically significant difference in rates of ectoparasite presence ($X^2 = 2.11$, d.f. = 3, $p = 0.549328$). Reproductive males had 62% ectoparasite presence and non-reproductive males had 82% ectoparasite presence. Female reproductive status was split into four separate categories; pregnant, lactating, post-lactating, and non-reproductive. Pregnant females had 24% ectoparasite presence, lactating females had 40% ectoparasite presence, and post-lactating and non-reproductive females both had 46% ectoparasite presence ($X^2 = 7.42$, d.f. = 7, $p = 0.38622$). Of the ectoparasites collected on *N. humeralis*, 82% were mites, 13% were cimicids, 0.15% were chewing lice, and 5% were unable to be identified.

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INTRODUCTION

The natural history of the northern myotis (*Myotis septentrionalis*), even in areas where it is widely distributed, is poorly understood. In Kansas, this species is listed as a species in need of conservation (SINC) and it was listed federally as threatened by the United States Fish and Wildlife Service in April 2015. The main cause of decline is thought to be due to White-nose Syndrome (WNS) caused by the fungus, *Pseudogymnoascus destructans*. Prior to my study, WNS was not observed within the state of Kansas, but recent surveys by the Kansas Department of Wildlife, Parks, and Tourism (KDWP&T) have confirmed the presence of WNS in Barber Cherokee, Comanche, Kiowa, Pratt, and Rooks counties in Kansas (Fig. 1).

To understand one aspect of the natural history of *M. septentrionalis*, I looked at ectoparasite presence and species. I did not limit my collection to only *M. septentrionalis*; I collected ectoparasites from the bycatch of Kansas bat species that I captured over the course of the 2016 and 2017 field seasons.

Many bat ectoparasites are still poorly known around the world (Ueshima 1972; Hopla et al. 1994; Ritzi 2004). An ectoparasite is defined as an organism that inhabits the skin, or outgrowths on the skin, of another organism (host) for varying periods, and might be detrimental to that organism (Hopla et al. 1994). I listed the ectoparasites typically associated with bats in the United States and Canada along with the bat species with which they were associated (Table 1; Jones et al. 1952; Sealander and Young 1955; Brennan and White 1960; Whitaker 1973; Whitaker and Wilson 1974; Whitaker and Loomis 1979; Dood and Kurta 1988; Dick et al. 2003; Reeves et al. 2007; Whitaker et al. 2007; Poissant and Broders 2008). Also, I listed whether the ectoparasites have been recorded in Kansas. All of these ectoparasites are known to feed on their host and can be detrimental to the health of the host if they are present in large numbers

(Whitaker and Wilson 1974; Herrin and Tipton 1975; Hudson et al. 2002; Sparks et al. 2003; Hudson et al. 2006; Lučan 2006; Bartonička and Gaisler 2007; Whitaker et al. 2007). The presence of ectoparasites can be quite taxing to the host and can result in behavioral changes to help the host attempt to rid itself of ectoparasites (ter Hofstede and Fenton 2005; Lučan 2006; Lourenço and Palmeirim 2007; Thrall et al. 2007). Some ectoparasites are species specific while others are more generalist and will parasitize a wide range of hosts (Krasnov et al. 2007). Below I summarize the life histories of some bat ectoparasites (primarily from Whitaker et al. 2009, except where cited).

Diptera: Nycteribiidae (Bat Flies)

Originally, this family was placed within the family Polyctenidae, as both groups are very similar, but later work separated out the two groups (Ritzi 2004). This family is divided into three subfamilies: Archinycteribiinae, Cyclopodiinae, and Nycteribiinae. The subfamilies Archinycteribiinae and Cyclopodiinae are located exclusively in the Western Hemisphere and are associated with pteropid bats. The remaining subfamily, Nycteribiinae, has a cosmopolitan distribution, and is associated mainly with the families Vespertilionidae and Rhinolophidae.

All members of this family are obligate, blood-sucking, specialized, true flies that are parasitic on bats. Although they are a true fly, all nycteribiids are completely wingless. They have a very spider-like appearance, and their head and legs originate from the dorsal thoracic surface. Like fleas, nycteribiids have several ctenidia (combs) that aid in the protection of joints and organs or might help to keep the organism from being brushed out of the fur.

Adult nycteribiids spend their entire lives on their bat host. The only exception is when a female leaves to lay a larva on the roost walls. Females lay fully developed larvae (prepupae)

that pupate immediately. Males never leave their host and try to mate with any females that they encounter.

Diptera: Streblidae (Bat Flies)

Streblids are exclusively ectoparasites of bats. This family is divided into five subfamilies: Nycteriboscinae, Ascodipterinae, Nycterophiliinae, Streblinae, and Trichobiinae. The Nycteriboscinae and the Ascodipterinae are limited to the Eastern Hemisphere, while the Nycterophiliinae, the Streblinae, and the Trichobiinae are limited to the Western Hemisphere.

All streblids are external parasites with the exception on the females in the genus *Ascodipteron*. These females embed themselves into the skin of bats, effectively becoming endoparasitic. Many streblids are winged, but there are several genera with vestigial wings. Even fully winged species, however, are weak flyers. Like the nycteribiids, females only leave the host to lay a single egg on the roost walls.

Siphonaptera (Fleas): Ischnopsyllidae

Adult fleas are hematophagous ectoparasites of birds and mammals that can cause problems such as anemia, dermatitis, hypersensitivity, and pathogen transmission (Hopla et al. 1994). Fleas are small, laterally compressed, wingless insects, usually with ctenidia (Whitaker et al. 2009; Bitam et al. 2010). The family Ischnopsyllidae feeds exclusively on the following bat families: Desmodontidae, Emballonuridae, Megadermatidae, Molossidae, Noctilionidae, Pteropodidae, Rhinolophidae, Rhinopomatidae, and Vespertilionidae (Marshall 1982; Ritzi 2004; Whitaker et al. 2009). Only the adults are parasitic, with a few exceptions.

Hemiptera (True Bugs): Cimicidae (Bed Bugs)

Most species of cimicid are obligate hematophages (Ritzi 2004). They live in bat roosts and bird nests, only coming out to feed. Many species of cimicid parasitize bats, swifts,

swallows, or humans, by taking advantage of their gregarious, or social lifestyles (Reinhardt and Siva-Jothy 2007). Many cimicids feed on bats, but some species feed on humans, such as *Cimex hemipterus* and C. *lectularius*. Cimicids are small, oval-shaped, dorsoventrally flattened with non-functioning wing pads, and piercing-sucking mouthparts (Jones and Jordan 1991). They do not have ctenidia or clasping tarsi that are often seen on permanent parasites and can survive long periods without a blood meal.

Cimex adjunctus is a species that is commonly parasitic on bats and is often confused with *C. lectularius* because they are very similar in appearance (Jones and Jordan 1991). *Cimex adjunctus* and *C. lectularius* are so similar that they can only be told apart by using a microscope. The fringe hairs along the pronotum are as long as, or longer than the width of the eyes on *C. adjunctus* (Fig. 2). On *C. lectularius* the fringe hairs along the pronotum and are shorter than the width of the eye (Jones and Jordan 1991).

On bats, cimicids are associate with hairless areas such as the wings, forearms, uropatagium, feet, and penis (Reinhardt and Siva-Jothy 2007). *Cimex adjunctus* is a free-living ectoparasite of bats, but in the absence of their preferred host will feed on humans. This typically is seen when bats roost in a home and are then removed leaving the cimicids behind (Jones and Jordan 1991). *Cimex adjunctus* will hide in the cracks and crevices in bat roosting areas and make repeated visits to the bat throughout the day. This same behavior can be seen in *C. lectularius* as well (Jones and Jordan 1991; Reinhardt and Siva-Jothy 2007).

Effects on the host include an immune response, causing discomfort; secondary infection; physiological changes in the host; and a change in the host's reproductive success (Reinhardt and Siva-Jothy 2007). Bats can rid themselves of these ectoparasites by grooming or changing roost

sites. Moving roost sites can be very effective for bats to rid themselves of these parasites, as eggs are laid and cemented to the roost and the bats are unlikely to carry eggs to a new roost.

Hemiptera (True Bugs): Polyctenidae (Bat Bugs)

Bats bugs are a rare group of hemipterans (Polyctenidae) that are permanent ectoparasites of bats (Ritzi 2004; Whitaker et al. 2009). There is only one genus of polyctenid, *Hesperoctenes*, thought to occur in the New World (Ueshima 1972; Whitaker et al. 2009). In the United States, only two species are known from California and Texas, *H. eumops* and *H. hermsi*, respectively. These were associated with *Eumops perotis* (*E. californicus*) and *Tadarida macrotis* (*T. molossa*), respectively (Ueshima 1972).

Polyctenids can be confused with cimicids but have a few key differences that separate them. They have a longer, thinner body, lack eyes, and have modified limbs for clinging onto the hair of bats. Unlike cimicids, they are obligate ectoparasites that can only go a short time without feeding and spend their entire life on their host (Ritzi 2004; Whitaker et al. 2009). Polyctenids also are viviparous, a trait unique among Hemiptera, so they do not need to leave the host to lay eggs.

Acarina (Mites, Ticks, and Chiggers): Argasidae and Ixodidae (Ticks)

The tick families Argasidae (soft-bodied ticks) and Ixodidae (hard-bodied ticks) both have been associated with bats, although argasids are more common. Many species of argasids are associated with birds, and some species associated with bats are even associated more commonly on birds. This could be due to ticks being habitat specific, rather than host specific. Thus, they are limited by their habitat, and will feed on anything with blood that uses that habitat.

Hard ticks and soft ticks both must have a blood meal each time they molt. For hard ticks, the time spent on the host can be considerable, as they need to stay attached for several days so that they can molt. Soft ticks can do several feedings in rapid succession as they only need to feed for a few minutes at a time. Female soft ticks can lay up to 500 eggs in the roost in between each feeding, and all stages live in the roost.

Acarina (Mites, Ticks, and Chiggers): Trombiculidae and Leeuwenhoekiidae (Chiggers)

"Chigger" is the term used to refer to the larval stage of a mite in the family Trombiculidae or the family Leeuwenhoekiidae. Many chiggers are parasites on vertebrates, while the free-living stages are predators of arthropods and their eggs, these stages are poorly known, however. As a result, classification of Trombiculidae and Leeuwenhoekiidae is based mostly on the larval chigger stage.

Chiggers, like ticks, are habitat restricted, and will feed on any vertebrate entering the area. These parasitic larvae serve as a method of dispersal, while the postlarval stages restrict a species to a habitat. This habitat restriction by these postlarval stages can limit the hosts available to larvae. Species associated with bats appear to be able to feed on other organisms, but they are restricted from other hosts by their habitat specializations. The species *Albeckia senase* is usually found on *Myotis velifer*, a primarily cave-roosting bat. In Kansas though, there was an instance where *A. senase* was recovered from the southern plains woodrat (*Neotoma micropus)*, that was found in a cave where bats were roosting.

Acarina (Mites, Ticks, and Chiggers): Cheyletidae (Mites)

Most cheyletid mites are free-living predators, but some could be parasitic on mammals (Whitaker et al. 2007; Whitaker et al. 2009). *Cheletonella vespertilionis* is known from a bat in Australia, and from a bat in Indiana. It also is known from the guano piles of a colony of

Eptesicus fuscus in Indiana, and the guano piles of *Myotis velifer* in Texas. Mites of this family are characterized by a large terminal claw, which is usually toothed, on the palpal tibia, each leg usually has two claws and an empodium (Whitaker et al. 2007).

Acarina (Mites, Ticks, and Chiggers): Chirodiscidae (Mites)

Chirodiscid mites are very similar to listrophorid mites, but chirodiscid mites have legs 1 and 2 highly modified for grasping hairs. Four North American genera, *Alabidocarpus*, *Dentocarpus*, *Olabidocarpus*, and *Schizocarpus*, formerly were included in the family Listrophoridae. The genera *Alabidocarpus*, *Dentocarpus*, and *Olabidocarpus* have all been associated with bats in the United States (Whitaker et al. 2007).

Acarina (Mites, Ticks, and Chiggers): Laelapidae (Mites)

Many laelapid mites are parasitic, but only a few are parasitic on bats. The four species known to be parasitic on bats are: *Notolaelaps novaguinea*, *Neolaelaps spinosa*, *N. vitzhumi*, and *N. palpispinosus* from bats in New Guinea, Asia, Australia, and oceanic islands. Other genera reported on bats, *Androlaelaps* and *Laelaps*, are found frequently on other hosts and are considered to be accidental infestations (Whitaker et al. 2007; Whitaker et al. 2009). Laelapid mites are identified by their jug-shaped epigynial plate, and an elongated peritreme (Whitaker et al. 2007).

Acarina (Mites, Ticks, and Chiggers): Macronyssidae (Mites)

Macronyssid mites are related closely to laelapid mites but can be differentiated by a pronounced ridge on the palpal trochanter (Whitaker et al. 2007). Many species are parasitic on bats, and feed on their blood or body fluids. Genera known to feed on bats include: *Chiroptonyssus*, *Cryptonyssus*, *Macronyssus*, *Ornithonyssus*, and *Steatonyssus* (Whitaker and Wilson 1974; Sparks et al. 2003; Whitaker et al. 2007).

Acarina (Mites, Ticks, and Chiggers): Myobiidae (Mites)

Myobiid mites are parasitic solely on mammals, and several species are parasites of bats (Whitaker et al. 2007; Whitaker et al. 2009). The first pair of legs are modified for grasping hairs, the chelicerae are minute, as are the palpi (Whitaker et al. 2007). These mites feed directly on the host's tissue fluids. All stages of life stay on the host, with adult females gluing their eggs to the host's hair. Genera that parasitize bats include: *Acanthophthirius*, *Ewingana*, *Phyllostomybia*, *Pteracarus*, and *Radfordia* (Whitaker and Wilson 1974; Whitaker et al. 2007).

Acarina (Mites, Ticks, and Chiggers): Pygmephoridae (Mites)

Within Pygmephoridae only the species *Pygmephorus mahunkai* has been recorded on bats, and thus far it is only known from the bat species *Myotis lucifugus*. Members of the genus *Pygmephorus*, however, are thought to be phoretic and not parasitic. Within this genus, individuals recorded are females usually that have an enlarged first pair of legs for grasping hair. This genus is also often associated with small mammals, especially insectivores, and are thought to feed on fungi in the soil or in the nests of mammals (Whitaker et al. 2007).

Acarina (Mites, Ticks, and Chiggers): Rosensteiniidae (Mites)

Rosensteiniids are associates of bats and their roosts, but they are not parasitic and commonly are not associated with bats. These mites could be abundant in guano and the roost, where they feed on feces or on smaller organisms. The genera *Chiroptoglyphus*, *Mydopholeus*, and *Nycteriglyphus* have all been associated with bats (Whitaker et al. 2007; Whitaker et al. 2009).

Acarina (Mites, Ticks, and Chiggers): Spinturnicidae (Mites)

Spinturnicid mites are all parasitic on bats. These mites have a reduced tritosternum, prominent legs, a stout body, and the peritreme is often ventral posteriorly. Most species crawl

on the wing and tail membranes, but the species *Paraspinturnix globosus* is known to live in the anus of some North American *Myotis* species. Genera associated with bats in the United States and Canada are: *Periglischrus*, and *Spinturnix* (Whitaker et al. 2007; Whitaker et al. 2009).

Phthiraptera (Lice): Anoplura (Sucking Lice) and Mallophaga (Chewing Lice)

Lice are flattened, wingless insects that are obligate hematophagous ectoparasites of birds and mammals (Hopla et al. 1994). Lice typically are divided into two suborders: Anoplura, the sucking lice, and Mallophaga, the chewing lice (Hopla et al. 1994; Ritzi 2004). The Mallophaga, however, have been further separated into three suborders: Amblycera, Ischnocera, and Rhyncophthirina. The Ischnocera and Rhyncophthirina mainly parasitize mammals, while the Amblycera mainly parasitize birds (Ritzi 2004). Sucking lice have a head that is narrower than its prothorax, while chewing lice have a head that is as wide as or wider than its prothorax (Johnson and Clayton 2003).

Kansas Bat Species

In Kansas 15 species of bats are known to occur, and there is one additional species that potentially might occur. All species are in the family Vespertilionidae unless otherwise indicated. The known occurrences are: *Antrozous pallidus*, pallid bat; *Corynorhinus townsendii*, Townsend's big-eared bat; *Eptesicus fuscus*, big brown bat; *Lasionycteris noctivagans*, silver-haired bat; *Lasiurus borealis*, eastern red bat; *Lasiurus cinereus*, hoary bat; *Myotis ciliolabrum*, western small-footed myotis; *Myotis grisescens*, gray myotis; *Myotis lucifugus*, little brown myotis; *Myotis septentrionalis*, the northern myotis; *Myotis velifer*, cave myotis; *Myotis yumanensis*, Yuma myotis; *Nycticeius humeralis*, evening bat; *Perimyotis subflavus*, tri-colored bat; and *Tadarida brasiliensis* (Molossidae), Brazilian free-tailed bat (Schmidt et al. 2019). The

species of questionable occurrence in Kansas is *Nyctinomops macrotis*, big free-tailed bat (Schmidt et al. 2019).

Myotis septentrionalis has historically occurred in Kansas, but recent records are lacking. As Kansas only recently has had cases of WNS (Fig. 1), it is critical to determine the status of the current *M. septentrionalis* population in Kansas. The overarching goal of the project, which began in 2015 and ended in 2019, was to try and determine where in Kansas these bats occur, characteristics of hibernacula, and characteristics of maternity colonies.

My main objective, while working on the project, was to identify which ectoparasites were present on Kansas bats. My second objective was to determine if there was any correlation between the presence of ectoparasites and age, reproductive status, and the sex of Kansas bats. Given the second objective, I formed the following three hypotheses. My first hypothesis was that juvenile bats had greater parasite presence than adults (McLean and Speakman 1997; Christe et al. 2003; Lučan 2006). My second hypothesis was that males would have a higher presence of ectoparasites then females (Lučan 2006). My third hypothesis was that reproductive female bats would have a higher presence of ectoparasites than non-reproductive bats (Lučan 2006).

METHODS

Bat Capture

All of the following methods fell within the approved Animal Care and Use Committee (IACUC) protocol (15-0002, Appendix 1). Bats were captured by using mist nets set across rivers and streams just before sunset (Carroll et al. 2002; Robbins et al. 2008). In the field season of 2016, I set mist nests in a total of 43 locations in three northcentral Kansas counties: Ellis, Rooks, and Russell. In the field season of 2017, I set mist nets in a total of 22 locations in eight Kansas counties: Butler, Ellis, Jewell, Marshall, Phillips, Rooks, Russell, and Trego (Fig. 3).

I chose netting localities by using a combination of historic localities for *M. septentrionalis* and acoustic monitoring by using an SM 3 Bat detector. I took historic localities recorded in the Kansas Mammal Atlas (Schmidt et al. 2019). I compiled historic localities as a combination of voucher specimens, visual observations, and literature observations. I set acoustic detectors near these historic localities and checked for the presence of *Myotis* species. If supposed *Myotis* calls were recorded, then I set mist nets in that location and in the surrounding areas.

I kept mist nets closed until sunset to avoid capturing birds. I checked mist nets every five to ten minutes for bats once they were opened. Frequent checking minimized stress on bats and ensured that bats did not chew holes in mist nets. I used single, double high, or triple high setups. I only used the triple high setup in the 2016 field season. Mist net lengths depended on the width of the waterway and were 6m, 9m, and 12m. In addition, in Russell County, I found bats in a 356m long, underground, man-made cave. Because it was difficult to set mist nets

outside either entrance of the cave, I collected bats by taking them from the walls, or by using butterfly nets.

After capture I placed each bat into a Dixie PerfecTouch 12-ounce, paper cup with a lid, and weighed the cup containing the bat by using a 50 g Pesola scale. The bats remained in a cup for a minimum of 30 minutes, but no longer than three hours. I did this to help ensure fecal sample collection for diet analysis. Following fecal collection, I checked each bat for age, sex, and reproductive status. I estimated age by shining a light through the wing membrane to determine if there was epiphyseal-diaphyseal fusion. If the joint was fused, I considered the individual to be an adult, and juvenile if not fused. I examined the bats to determine the sex based on the external genitalia. I determined males to be reproductive or non-reproductive based on the descended or non-descended, testes, respectfully. I placed females into one of four reproductive categories: pregnant, lactating, post-lactating, or non-reproductive. For each bat, I recorded in millimeters the following: ear length, tragus length, forearm length, body length, tail length, and hindfoot length. Finally, I examined each bat for ectoparasites, banded them and released them.

Ectoparasite Collection

I used a modification of Whitaker's method for ectoparasite collection (Whitaker et al. 2009). Collecting ectoparasites was done by two people and consisted of colleague holding a bat spread-winged for inspection, over a large-mouth plastic jar that contained 60% ethanol, while visual inspection and brushing was done. I visually inspected each bat initially for large ectoparasites, such as cimicids and ticks. I removed these large ectoparasites by using soft-tipped forceps and placed them in the 60% ethanol. Afterwards, I used a soft toothbrush to brush

the body and wings, both dorsally and ventrally so that smaller ectoparasites, such as mites, fell into the ethanol. I rinsed the toothbrush in ethanol between uses (Ritzi 2004; Whitaker et al. 2009). I brushed bats thoroughly until ectoparasites were no longer visible to the naked eye. I then banded each bat with an identification number and released onto the nearest tree away from the mist nets

Slide Preparation

I followed the techniques and instructions included with the insect slide mounting kit purchased through the BioQuip website (BioQuip 2001). I placed mites in a solution of 75% ethanol for 10 minutes, moved them to an 85% solution for another 10 minutes, and then placed them in a 95% ethanol solution for 10 minutes. This made the mites miscible (forming a homogenous mixture when added together) with the Euparal mounting solution. Finally, I placed individuals in one to two drops of Euparal mounting solution and positioned them ventral side up by using micro tools under a microscope. Once positioned I placed a cover slip on the slide and left it to dry on a slide heater. Cimicids were too large to place onto a slide, so I identified them under a dissecting microscope. I identified ticks under a dissecting microscope because they were soft-bodied and placing them onto a slide would have damaged identifiable characteristics. I deposited all 90 prepared slides with the Sternberg Museum of Natural History's entomology collection.

Ectoparasite Identification

I identified ectoparasites by using a combination of mite, tick, and cimicid dichotomous keys (Keegan 1951; Furman and Catts 1982). I identified mites by using a compound light microscope and ticks and cimicids by using a dissecting microscope.

Statistical Analysis

For my statistical analysis I used Pearson's chi-squared test of independence (Pearson 1900). I used this test because it assesses whether unpaired observations on two variables, expressed in a contingency table, are independent of each other. This allowed me to compare ectoparasite presence between age, reproductive status, and sex for each bat species captured.

RESULTS

Bat Capture

I captured bats beginning in early May and continuing until early October in 2016 and 2017 (Table 2). Between the 2016 and 2017 field seasons, I mist netted for 104 nights over a total of 248 hours (Table 2). The species of bats I captured: *E. fuscus* (N = 64), *L. borealis* (N = 10), *L. cinereus* (N = 5), *M. septentrionalis* (N =7), *N. humeralis* (N = 317), and *P. subflavus* (N = 80) (Table 3).

Ectoparasite Identification

I collected the following ectoparasites: mites in the families Laelapidae and Spinturnicidae; cimicids in the family Cimicidae; ticks in the family Argasidae, and a chewing louse (Fig. 4) in the order Mallophaga (Table 3). These ectoparasites were distributed across six species of bats (Table 3).

I identified mites in the family Laelapidae as the genus *Haemogamasus*, which previously has not been documented in Kansas. It has been documented in many parts of the United States, but it mostly has been limited to the coasts, with a single occurrence documented in Oklahoma (Whitaker and Wilson 1974). I found the laelapid mites in Ellis, Jewell, and Rooks counties; the spinturnicid mites in Ellis, Jewell, and Rooks counties; the cimicids in Ellis, Jewell, and Rooks counties; the ticks in Ellis County; and the louse in Ellis County. I did not find ectoparasites on bats in Coffey, Lyon, Marshall, Osage, Osborne, Phillips, Republic, Russell, Trego, or Washington counties. *Nycticeius humeralis*, however, was the only species captured in high enough numbers to evaluate statistically. This was somewhat unexpected because there has not been that many recent records of *N. humeralis* in northcentral Kansas. *Eptesicus fuscus* was

expected to be captured in greater numbers because they have been recorded in higher numbers in northcentral Kansas (Schmidt et al. 2019).

I collected ectoparasites from each captured bat species listed below. From *Eptesicus fuscus*, I collected 3 mites in the family Laelapidae and 3 mites in the family Spinturnicidae; 5 cimicids in the family Cimicidae; 3 ticks in the family Argasidae; and 4 individuals that I was unable to identify (Table 3). From *Lasiurus borealis*, I collected no ectoparasites (Table 3). From *Lasiurus cinereus*, I collected 3 mites in the family Spinturnicidae (Table 3). From *Myotis septentrionalis*, I only collected one unidentifiable mite. From *Nycticeius humeralis*, I collected 492 mites in the family Laelapidae and 67 mites in the family Spinturnicidae, 89 cimicids in the family Cimicidae, a single louse in the order Mallophaga, and 35 individuals that I was unable to identify (Table 3). Finally, I collected no ectoparasites from *Perimyotis subflavus* (Table 3).

Statistical Analysis

I used a chi-square test of independence to determine significance for the following comparisons. I compared the presence of ectoparasites between adult and juvenile *N. humeralis*. Juvenile *N. humeralis* had a higher presence of ectoparasites than adults ($X^2 = 47.38$, d.f. = 3, $p = 0.00001$, Table 4). I compared the presence of ectoparasites between male and female *N. humeralis*. Males had higher presences of ectoparasites than females ($X^2 = 15.03$, d.f. = 3, $p = 0.0018$, Table 4). I compared the presence of ectoparasites between reproductive and non-reproductive male *N. humeralis* and found no significant difference ($X^2 = 2.11$, d.f. = 3, $p = 0.55$, Table 4). When comparing the reproductive status of *N. humeralis* females, I split them into four separate categories: pregnant, lactating, post-lactating, and non-reproductive. Similar to males, there was no significant difference in ectoparasite presence between the female reproductive stages ($X^2 = 7.42$, d.f. = 7, $p = 0.39$, Table 4).

DISSCUSSION

The first objective of my project was to identify which ectoparasites were present on captured bats. I was able to meet my first objective by collecting ectoparasites from all bats captured over the two field seasons. For my first objective, however, I was only able to identify larger ectoparasites, such as cimicids and large mites, on the bats that were captured. I was unable to collect smaller ectoparasites, such as follicle mites, because my chosen method of collection was biased towards larger organisms. The best way to collect and identify smaller ectoparasites would have been to euthanize the bat, and then examine it under a microscope (Whitaker et al. 2009), which I could not do because of the overall objectives of the project. Furthermore, it is very time consuming, and is not ideal for threatened or endangered species (Whitaker et al. 2009).

The second objective of my project was to determine if there was any relationship between the presence of ectoparasites and age, reproductive status, and the sex of each bat species captured. I was able to meet my second objective by performing a chi-square test of independence to determine if there was any relationship between age, reproductive status, or sex, and the presence of ectoparasites. For my second objective, there were statistically significant differences between adult and juvenile *N. humeralis*, as well as between males and females. There was no statistically significant difference between different reproductive statuses for male and female *N. humeralis*.

My first hypothesis was that juvenile bats had greater parasite presence than adults (Christe et al. 2003; Lučan 2006). Due to a small sample size, I was only able to statistically analyze ectoparasites from *N. humeralis*. Juveniles had a higher presence of ectoparasites than adults did. This might be due to juveniles being restricted to their

roosts until they are able to fly, making them more susceptible to ectoparasites (McLean and Speakman 1997; Christe et al. 2000, 2003). Juveniles also might not be able to groom themselves as well as adults (Christe et al. 2003).

My second hypothesis was that males would have a higher presence of ectoparasites than females in *N. humeralis*. Males had a higher presence of ectoparasites than the females did. Males might have more ectoparasites due to their promiscuous nature. Males tend to try and mate with as many females as possible, thus, increasing their chance to come into contact with ectoparasites (Webber et al. 2015). Other bat species might not have had as high of levels of ectoparasites because of very low capture rates not being truly representative of the populations.

My third hypothesis was that reproductive female *N. humeralis* would have a higher presence of ectoparasites than non-reproductive bats (Lučan 2006). When looking at reproductive status there was no significant difference in ectoparasite presence. This might be because there was not a large enough sample (126 reproductive individuals, split into four reproductive categories), or because there is no relationship between ectoparasite presence and reproductive status. Just looking at the numbers, however, nonreproductive males had a higher presence of ectoparasites, as did the nonreproductive females.

There have been many studies looking at age, reproductive status, sex, and the presence of ectoparasites on bats, but the results have been contradictory. Reproductive females had a higher presence of ectoparasites than males did, and juveniles had a higher presence of ectoparasites than adults did for the species *Myotis blythii*, *M. daubentonii*, *M. emarginatus*, *M. myotis*, *M. mystacinus*, *M. nattereri*, *Nyctalus noctula*, *Pipistrellus*

pipistrellus, and *Plecotus auritus* (Christe et al. 2000, 2003; Zahn and Rupp 2004; Lučan 2006; Christe et al. 2007). However, adult *Eptesicus fuscus* in Colorado had a higher presence of ectoparasites than juveniles did, and lactating females had the highest presence of ectoparasites (Pearce and O'Shea 2007). Pregnant female *Miniopterus schreibersii* had the highest presence of ectoparasites in temperate-zone caves (Lourenço and Palmeirim 2008). These results most likely conflict due to numerous factors. Bat species and ectoparasite species both play a role in ectoparasite presence. Each species has its own life history, and many ectoparasites have a very poorly understood life history. Roosting sites, temperature, time of year, and geographic region all likely factor in as well (McLean and Speakman 1997; Christe et al. 2000, 2003, 2007; Zahn and Rupp 2004; Lučan 2006; Pearce and O'Shea 2007; Lourenço and Palmeirim 2008).

Individual populations of bats and ectoparasites need to be studied because relationships might not be the same even across the same species. For example, 80 *Perimyotis subflavus* were captured (all were in the cave except one), but no ectoparasites were collected, even though ectoparasites have been collected from this species in past studies (Table 1; Jones et al. 1952; Whitaker and Wilson 1974; Whitaker et al. 2007). Ectoparasites could have been overlooked during collection or could have been missed by the sampling technique used. Possibly, lack of ectoparasites might be due to individuals roosting in small groups and keeping themselves very clean. This cave is man-made, and is roughly 200 years old, and could have been used for food storage in the past where pesticides were used to help keep food free of insects. This is another potential explanation for the lack of ectoparasites found on *P. subflavus*. Another reason might be that the cave is too cold or sterile for ectoparasites to maintain themselves in the roosts

while the bats are away. These differences could also be due to the time of the year I captured bats and checked for ectoparasites. Ectoparasite loads are known to fluctuate seasonally, and the inability to net all locations at the same time could have skewed the results (Lučan 2006; Bartonička and Gaisler 2007; Lourenço and Palmeirim 2008).

The single louse found is a tentative identification. The specimen did not transfer well to the slide, and key identifiable features were hard to see. It was also tentative because it was the only specimen found on 482 bats. Many of these bat species roost under the bark of trees, this louse might have been picked up accidentally from a bird's nest. Birds are common hosts of lice, and their nests can have a large number of lice found in them as well (Boyd 1951; Dunn 2005). Plausibly the bat could have picked up a louse by accident when roosting in or near a bird's nest.

Given that white-nose syndrome continues to spread in bats, we need to learn about other factors that might weaken bats and potentially make them more susceptible to the disease. This is particularly critical in places such as Kansas where the fungus only recently has been found. Understanding the impact ectoparasites can have on their bat hosts could potentially help with their management and successful recovery for threatened and endangered species, many of which are declining from white-nose syndrome.

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TABLES

Table 1. -- Known ectoparasite species associated with bats in the United States and Canada. *Antrozous pallidus* (ANPA); *Choeronycteris mexicana* (CHME); *Corynorhinus rafinesquii* (CORA); *C. townsendii* (COTO); *Eptesicus fuscus* (EPFU); *Euderma maculatum* (EUMA); *Eumops perotis* (EUPE); *Lasionycteris noctivagans* (LANO); *Lasiurus borealis* (LABO); *L. cinereus* (LACI); *L. intermedius* (LAIN); *Mormoops megalophylla* (MOME); *Myotis austroriparius* (MYAU); *M. californicus* (MYCA); *M. evotis* (MYEV); *M. grisescens* (MYGR); *M. keenii* (MYKE); *M. leibii* (MYLE); *M. lucifugus* (MYLU); *M. occultus* (MYOC); *M. septentrionalis* (MYSE); *M. sodalis* (MYSO); *M. thysanodes* (MYTH); *M. velifer* (MYVE); *M. volans* (MYVO); *M. yumanensis* (MYYU); *Nycticeius humeralis* (NYHU); *Nyctinomops femorosaccus* (NYFE); *N. macrotis* (NYMA); *Parastrellus hesperus* (PAHE); *Perimyotis subflavus* (PESU); and *Tadarida brasiliensis* (TABR). Y: yes; N: no

Order	**Family**	**Ectoparasite Species**	**Synonym(s)**	**Bat Species**	**Found on Kansas Bats**
Diptera	Nycteribiidae	Unidentified		ANPA, CORA, MYTH	N
		Basilia forcipata		MYOC, MYVO	N
	Streblidae	Unidentified		CHME, CORA	N
		Trichobius corynorhini		CORA	N
Siphonaptera	Ischnopsyllidae	*Myodopsylla sp.*		MYYU	N
		M. gentilis		MYOC	N
		M. insignis		EPFU, MYLU, MYSE	N
Hemiptera	Cimicidae	*Cimex sp.*		EPFU, PAHE	N
		C. adjunctus		EPFU, LANO. MYKE, MYLU, MYSE, NYHU	Y; EPFU, NYHU
		C. pilosellus		MYOC	N
	Polyctenidae	*Hesperoctenes eumops*		EUPE	N
Acarina	Argasidae	*Cryptonysuss desultorius*		MYLE	N
		Ornithodorus sp.		MYOC	N

		O. kelleyi		EPFU, MYLE, MYLU, MYSE	N
	Ixodidae	*Dermacentor sp.*		ANPA, PAHE	N
	Glycyphagidae	*Glycyphagus hypudaei*		MYOC	N
	Anoetidae	Undescribed *sp.*		EPFU	Y; EPFU
	Cheyletidae	*Cheletonella vespertilionis*		EPFU	N
		Cheyletus cacahuamilpensis		MYVE	N
	Chirodiscidae	*Alabidocarpus sp.*		EPFU	N
	Chirodiscidae	*A. calcaratus*	*A. longipilus*	MYCA, MYLU, MYOC, MYVO, MYYU	N
		A. eptesicus		EPFU, MYOC	N
		Dentocarpus macrotrichus		TABR	N
		Olabidocarpus whitakeri		MYAU, MYGR, MYSE	N
	Demoicidae	*Demodex*		MYSE	N
	Dermanyssidae	*Dermanyssus gallinae*	*D. evotomydis*	MYVE	N
	Glycophagidae	*Glycyphagus hypudaei*		MYOC	N
	Laelapidae	*Androlaelaps casalis*		TABR, MYLE, MYSE	N
		A. fahrenholzi	*Atricholaelaps sigmodoni; Haemolaelaps scalopi; Laelaps californicus; L. glassgowi scalopi; L. stegemani; L. virginianus; Liponyssus setiger*	COTO, EPFU, MYGR, NYHU, PESU, TABR	Y; EPFU, MYGR, TABR

		Eubrachylaelaps debilis		ANPA	N
		Haemogamasus ambulans	*H. alaskensis; H. sternalis; H. twitchelli*	MYCA, MYLU	N
		Ichoronyssus sp.		CORA, MYCA, MYTH	N
		Ischyropoda armatus		MYCA	N
		Laelaps alaskensis		MYLU	N
	Listrophoridae	*Dentocarpus macrotrichus*		TABR	N
		Listrophorus mexicanus		ANPA	N
		Olabidocarpus americanus		LAIN	N
		O. lawrencei		TABR	N
		O. whitakeri		MYAU, MYKE, MYSE	N
	Macronyssidae	*Chiroptonyssus haematophagus*		EUPE, MYOC	N
		C. robustipes	*C. texensis*	EPFU, LACI, MOME, MYCA, MYLU, MYOC, MYVE, TABR	Y; TABR
		C. venezolanus		NYFE, NYMA, PAHE	N
		Cryptonyssus sp.		MYOC	N
		C. desultorius		EPFU, EUMA, MYCA, MYOC, MYSO, MYVE, MYVO, MYYU, PAHE	N

		C. flexus		LANO, MYLU	N
		Macronyssus crosbyi	*Ichoronyssus brittanicus; I. quadridentatus*	CORA, COTO, EPFU, MYAU, MYCA, MYCI, MYEV, MYKE, MYLE, MYLU, MYOC, MYSE, MYSO, MYTH, MYVE, MYYU, NYHU, PESU	Y; MYSE
		M. jonesi	*Ichoronyssus; Macronyssus*	EPFU, MYAU, MYGR, MYVE	Y; EPFU, MYGR, MYVE
		M. longisetosus		COTO, MYVE	N
		M. macrodactylus		LANO	N
		M. unidens		COTO, EPFU, LANO, MYLU, MYVE, PESU	Y; COTO, EPFU, MYLU, MYVE
		Ornithonyssus sylviarum	*Liponyssus americanus; L. pacificus; O. banksi*	EPFU, MYVE	N
		Steatonyssus sp.		ANPA	
		S. antrozoi		ANPA, COTO, EPFU, MYYU	N
		S. ceratognathus		EPFU, LABO, MYLU, MYSE,	Y; NYHU

				MYSO, NYHU, PESU, TABR	
		S. emarginatus		MYYU, PAHE	N
		S. furmani		LABO, LACI	Y; LABO
		S. joaquimi		EPFU, MYLU	N
		S. occidentalis		CO sp., EPFU, LABO, MYLU, MYSO, MYVE, PAHE, PESU, TABR	Y; CO sp., EPFU, LABO, MYLU, MYVE, TABR
		S. radovsky		LAIN	N
	Myobiidae	*Acanthophthirius sp.*		LABO, LANO, MYKE, MYLU, MYSE, MYSO	N
		A. caudata	*Myobia canadensis*	EPFU, MYLU, MYVE, PESU	N
		A. gracilis		MYSE, MYVO	N
		A. lasiurus		LABO, LACI	N
		A. lucifugus		MYLU, MYOC, MYSO	N
		A. nycticeius		NYHU	N
		A. oregonensis		PAHE	N
		A. steatocaudatus		LANO	N
		Ewingana inaequalis		ANPA, TABR	N
		E. longa		TABR	N
		Pteracarus aculeus		EPFU	N

		P. chalinolobus		COTO, EPFU, LACI, LAIN, MYCI, MYTH, MYVO	N
		P. completus		LABO, LACI, EPFU	N
		P. elegans		COTO	N
		P. minutus		MYOC, MYVO, PESU	N
		P. robustus		ANPA	N
		Radfordia floridensis		TABR	N
	Pygmephoridae	*Pygmephorus mahunkai*		MYLU	N
	Rosensteiniidae	*Chirptoglyphus americanus*		CORA, MYLU, MYVE	N
		Nycteriglyphus bifolium		TABR	N
		N. fuscus		EPFU	N
		N. pennsylvanicus		EPFU	N
		N. texanus		COTO, MYVE, MYYU, TABR	N
	Spinturnicidae	*Paraspinturnix globosus*		MYSO	N
		Periglischrus strandtmanni		MOME	N
		Spinturnix sp.		ANPA, CHME, EPFU, MYCA, MYTH, MYYU	N
		Spinturnix americanus	*Pteroptus echinipes; S. carloshoffmanni; S. grossus, S. iowae*	ANPA, EPFU, MYAU, MYCI, MYEV, MYGR,	Y; EPFU, MYGR, MYLU, MYVE, MYYU, PESU,

				MYKE, MYLE, MYLU, MYOC, MYSE, MYSO, MYTH, MYVE, MYVO, MYYU, PESU, TABR	TABR
		S. bakeri	*S. americana; S. americanus; S. echinipes (?)*	EPFU, MYVO	Y; EPFU
		S. banksi		MYGR, MYVE	Y; MYGR, MYVE
		S. carloshoffmanni		MYVE	N
		S. globosus		MYGR, MYLU, MYSO, MYVE, MYVO	Y; MYGR, MYVE
		S. iowae		MYGR, MYSO, PESU	N
		S. myoti	*Pteroptus grossus*	Bat	N
		S. orri	*S. americanus*	ANPA	N
	Trombiculidae	*Euschoengastia hamiltoni*		EPFU, MYLE	N
		E. pipistrelli		EPFU, MYAU, MYGR, MYKE, MYLU, MYSE, PESU	Y; MYSE
		Eutrombicula alfreddugesi		MYSE	N
		Leptotrombidium myotis		EPFU, MYKE.	N

				MYLE, MYLU, MYOC, MYSE, MYVO	
		Neotromibcula microti		EPFU	N
		Parasecia gurneyi		EPFU	N
		Trombicula alfreddugesi	*Eustrombicula alfreddugesi*	EPFU, LABO, MYKE	N
		T. batatas	*E. batatas*	LABO	N
		T. gurneyi		NYHU	N
		T. myotis		MYGR	N
		Whartonia senase		EPFU	N
	Dermestidae	*Dermestes sp.*		EPFU	Y; EPFU

Table 2. -- Number of nights and hours spent mist netting for each month of the 2016 and the 2017 field seasons, with the number and species of bats captured. Except for a single individual, all PESU were captured at the man-made tunnel. *Eptesicus fuscus* (EPFU); *Lasiurus borealis* (LABO); *Lasiurus cinereus* (LACI); *Myotis septentrionalis* (MYSE); *Nycticeius humeralis* (NYHU); and *Perimyotis subflavus* (PESU).

Month/Year	**Number of nights and hours spent mist netting**	**Bat species and numbers captured**
May 2016	7 nights; 17 hours, 6 minutes	EPFU: 4 NYHU: 24
June 2016	20 nights; 49 hours, 33 minutes	EPFU: 4 LABO: 3 LACI: 1 MYSE: 2 NYHU: 64 (4 recaptured)
July 2016	16 nights; 35 hours, 48 minutes	EPFU: 38 (1 recaptured) LABO: 4 LACI: 5 MYSE: 1 NYHU: 88 (1 recaptured)
August 2016	8 nights; 16 hours, 27 minutes	EPFU: 2 LABO: 2 NYHU: 24 (1 recaptured)
September 2016	5 nights; 14 hours, 3 minutes	EPFU: 1 NYHU: 6 PESU: 21 (1 recaptured)
October 2016	3 nights; 12 hours, 30 minutes	MYSE: 1 PESU: 36 (8 recaptured)
May 2017	2 nights; 6 hours, 32 minutes	EPFU: 3 MYSE: 1 NYHU: 7 (1 recaptured)
June 2017	10 nights; 22 hours, 32 minutes	EPFU: 8 LABO: 2 MYSE: 2 NYHU: 46 (10 recaptured) PESU: 1
July 2017	14 nights; 29 hours, 55 minutes	EPFU: 1 NYHU: 30
August 2017	12 nights; 30 hours	EPFU: 2 LABO: 3 LACI: 1 NYHU: 49 (2 recaptured) PESU: 3
September 2017	5 nights; 9 hours, 49 minutes	EPFU: 4 (2 recaptured) NYHU: 1 PESU: 11 (2 recaptured)

October 2017	2 nights; 3 hours, 45 minutes	PESU: 53 (19 recaptured)

Table 3. -- Ectoparasites associated with select bat species in northcentral Kansas during the 2016 and 2017 field seasons. These are the total numbers of each type of ectoparasite associated with each species, not ectoparasites per individual. *Eptesicus fuscus* (EPFU); *Lasiurus borealis* (LABO); *Lasiurus cinereus* (LACI); *Myotis septentrionalis* (MYSE); *Nycticeius humeralis* (NYHU); and *Perimyotis subflavus* (PESU).

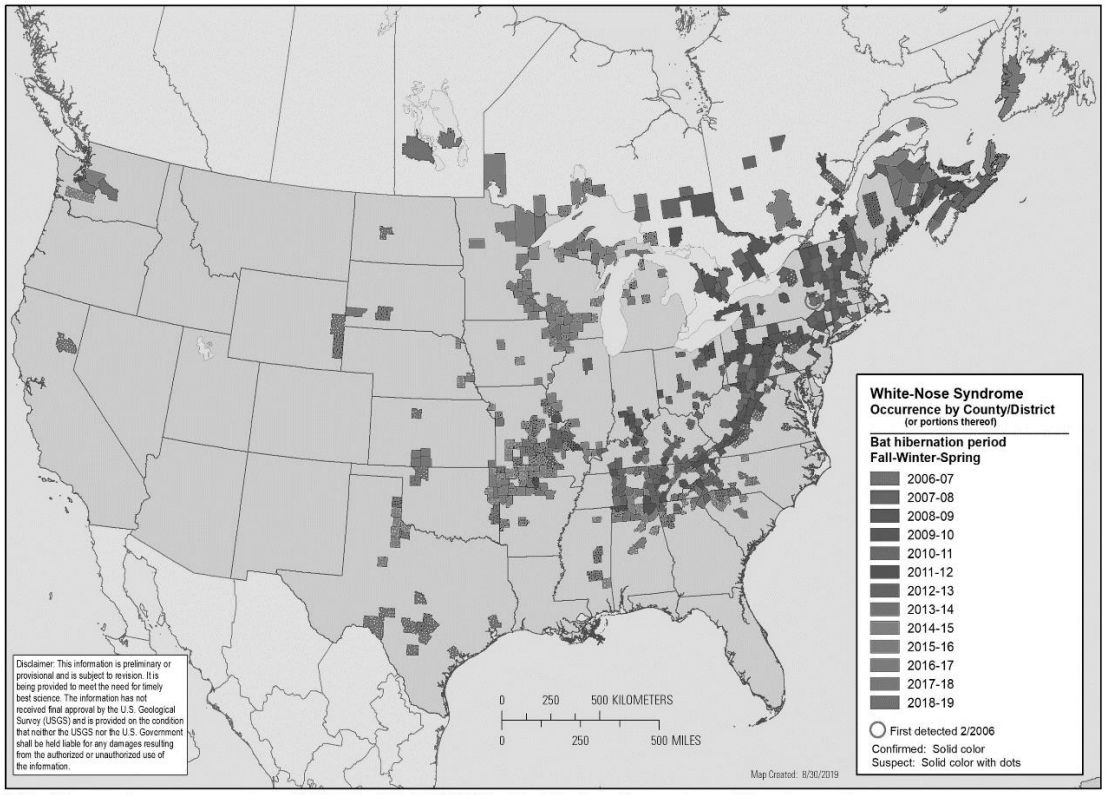
Species	**Number of bats**	**Bats with ecto-parasites**	**Laelapidae**	**Spinturnicidae**	**Chewing lice**	**Cimicids**	**Ticks**	**Un-known**
EPFU	64	8	3	3	0	5	3	4
LABO	10	0	0	0	0	0	0	0
LACI	5	2	3	0	0	0	0	0
MYSE	7	0	0	0	0	0	0	1
NYHU	317	143	492	67	1	89	0	35
PESU	80	0	0	0	0	0	0	0

Table 4. -- Statistical analyses showing the X^2 values and p-values between compared groups of *Nycticeius humeralis*. The number of bats with and without ectoparasites are shown for each category. The percentages of bats with and without ectoparasites for each group are also shown.

Categories	**X^2 value**	**P-value**	**Number of bats with ectoparasites**	**Number of bats without ectoparasites**	**Percentage with ectoparasites**	**Percentage without ectoparasites**
Adults	47.38	0.00001	74	149	33%	67%
Juveniles			69	22	76%	24%
Males	15.03	0.0018	33	13	72%	28%
Females			111	160	41%	59%
Reproductive males	2.11	0.5493	15	9	62%	38%
Nonreproductive males			18	4	82%	18%
Pregnant females	7.42	0.3862	12	37	24%	76%
Lactating females			21	32	40%	60%
Postlactating females			19	22	46%	54%
Nonreproductive females			56	69	46%	54%

FIGURES

Fig. 1. Map of the spread of white-nose syndrome (WNS), presented by the United States Fish and Wildlife Service (USFWS).



Citation: White-nose syndrome occurrence map - by year (2019). Data Last Updated: 8/30/2019. Available at: https://www.whitenosesyndrome.org/static-page/wns-spread-maps.

Fig. 2. Photograph showing how to differentiate between *Cimex lectularius* and *C. adjunctus*. The fringe hairs along the pronotum are as long as, or longer, than the width of the eyes on *C. adjunctus*. On *C. lectularius* the fringe hairs along the pronotum are shorter than the width of the eye (Jones and Jordan 1991). Photo courtesy of N. T. Gallagher.

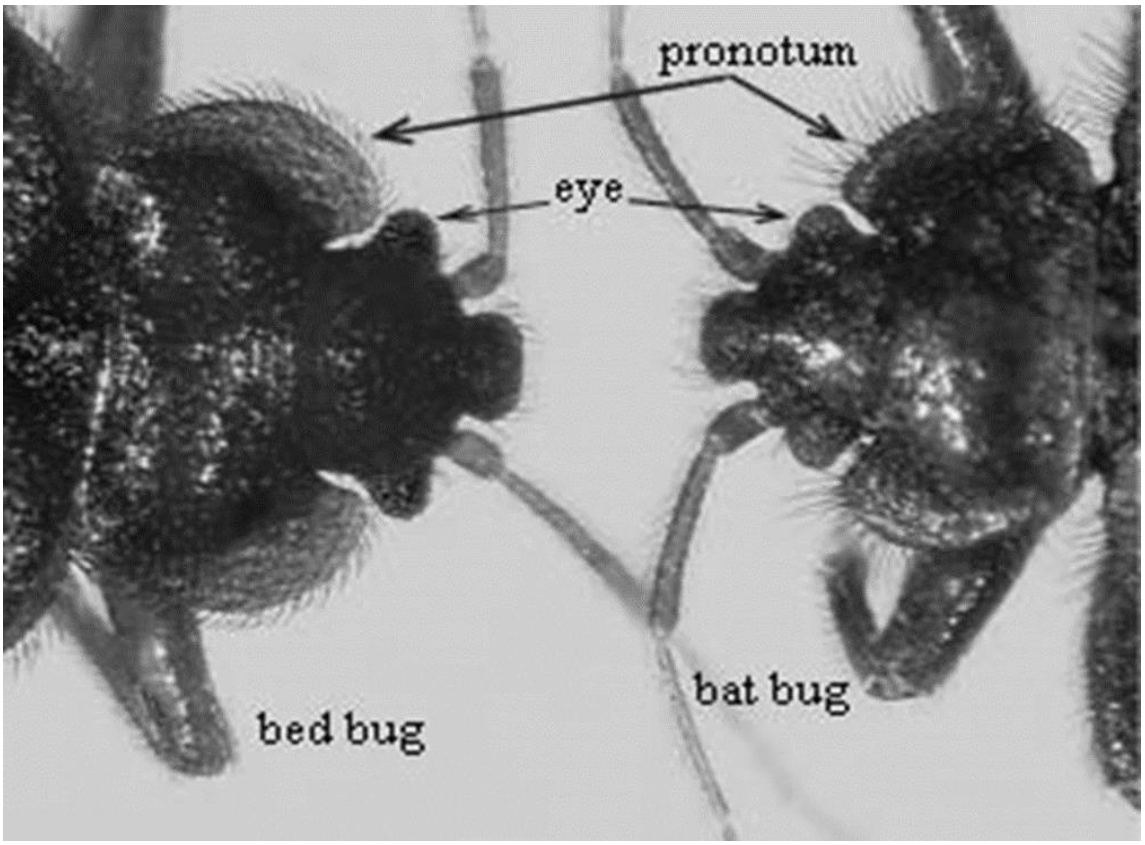


Fig. 3. Map of Kansas depicting the locations that were mist netted during the 2016 and 2017 field seasons.

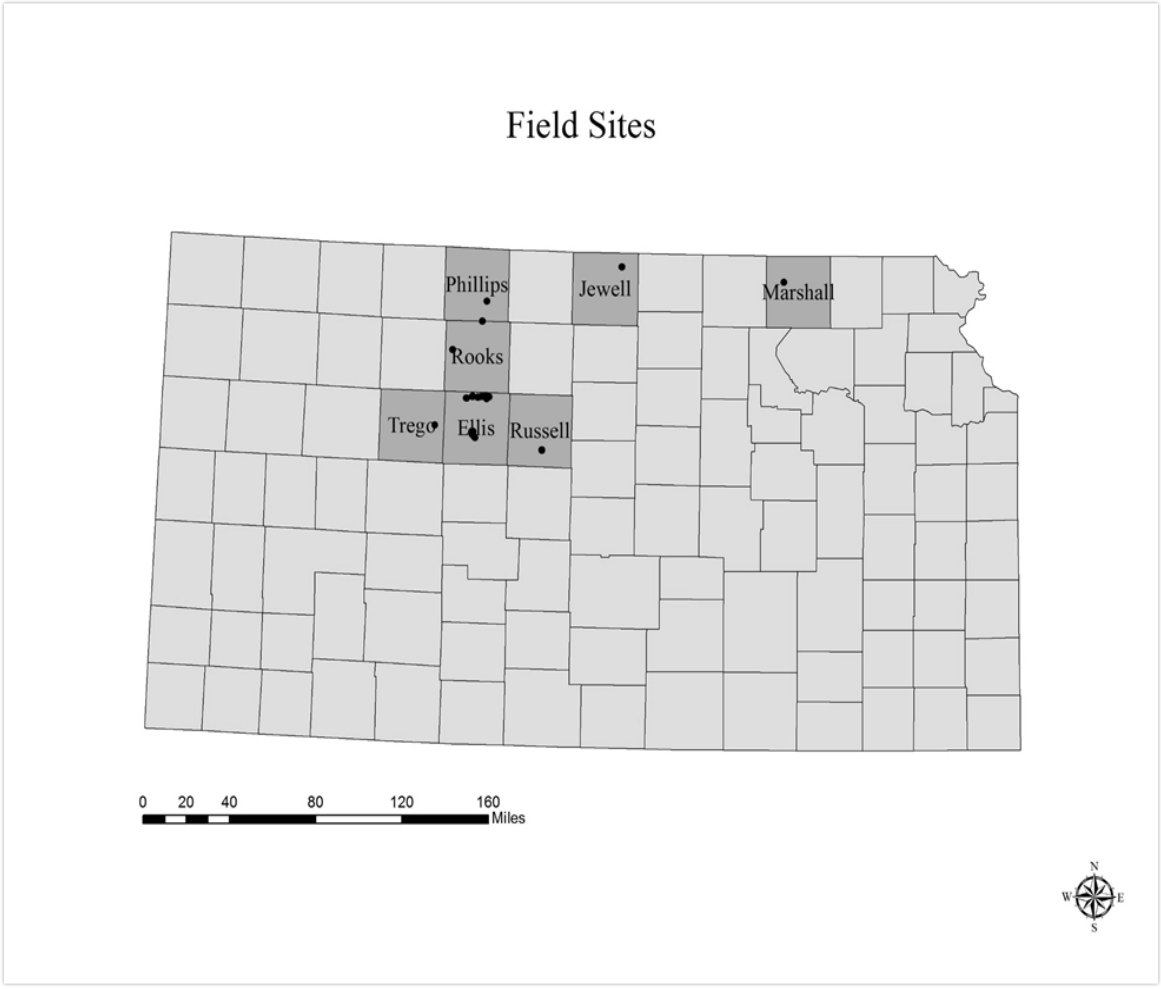
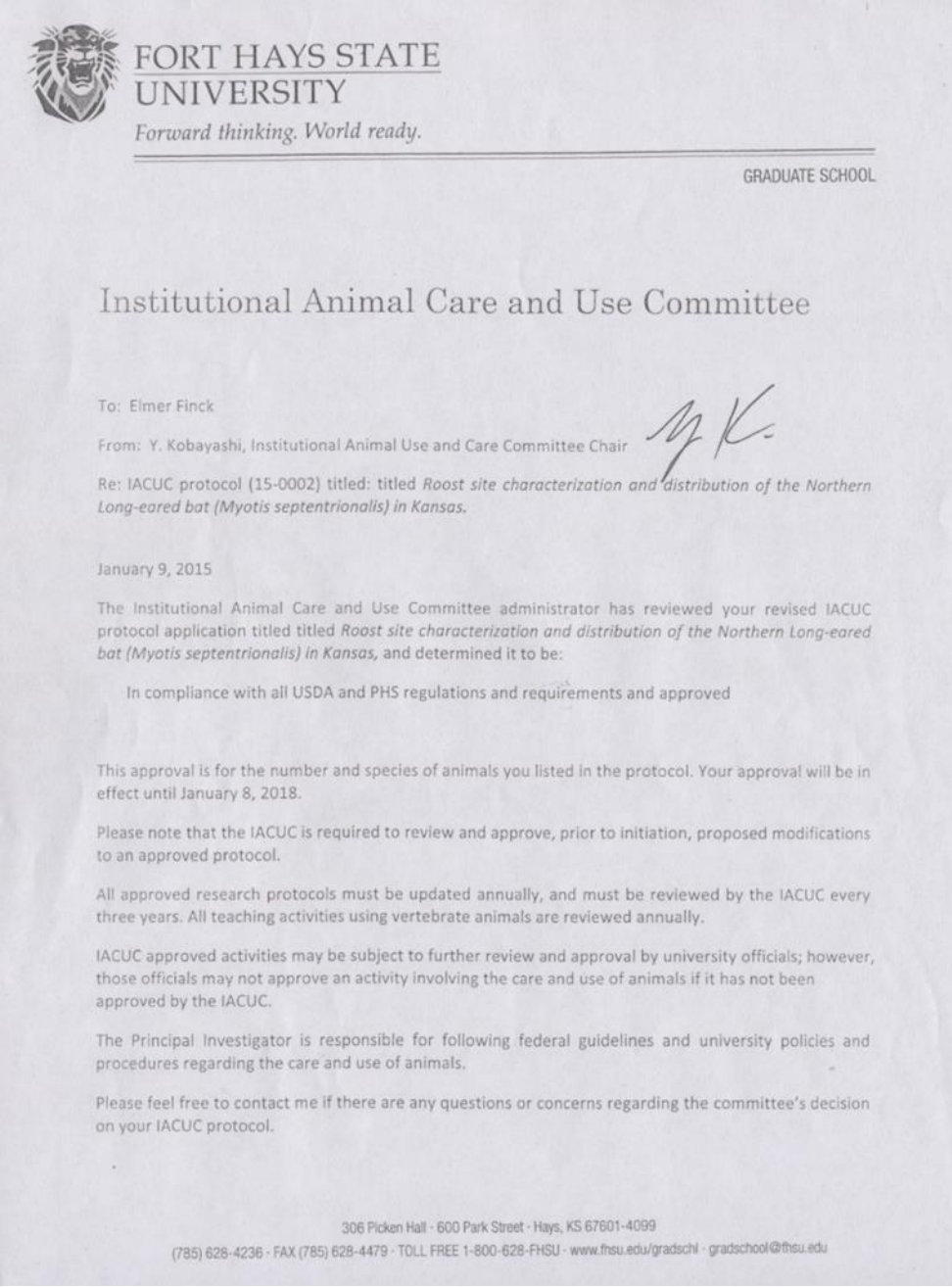


Fig. 4. Photograph of the chewing louse from Ellis County, Kansas.

APPENDICES

Appendix 1. – Project approval by the Fort Hays State University Institutional Animal Care and Use Committee protocol number 15-0002.



FORT HAYS STATE UNIVERSITY

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GRADUATE SCHOOL

Institutional Animal Care and Use Committee

To: Elmer Finck

From: Y. Kobayashi, Institutional Animal Use and Care Committee Chair

Re: IACUC protocol (15-0002) titled: titled *Roost site characterization and distribution of the Northern Long-eared bat (Myotis septentrionalis) in Kansas.*

January 9, 2015

The Institutional Animal Care and Use Committee administrator has reviewed your revised IACUC protocol application titled titled *Roost site characterization and distribution of the Northern Long-eared bat (Myotis septentrionalis) in Kansas,* and determined it to be:

In compliance with all USDA and PHS regulations and requirements and approved

This approval is for the number and species of animals you listed in the protocol. Your approval will be in effect until January 8, 2018.

Please note that the IACUC is required to review and approve, prior to initiation, proposed modifications to an approved protocol.

All approved research protocols must be updated annually, and must be reviewed by the IACUC every three years. All teaching activities using vertebrate animals are reviewed annually.

IACUC approved activities may be subject to further review and approval by university officials; however, those officials may not approve an activity involving the care and use of animals if it has not been approved by the IACUC.

The Principal Investigator is responsible for following federal guidelines and university policies and procedures regarding the care and use of animals.

Please feel free to contact me if there are any questions or concerns regarding the committee's decision on your IACUC protocol.

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